

ETHOLOGY OF *CEROTAINIA ALBIPILOSA* CURRAN
(DIPTERA: ASILIDAE) IN MARYLAND:
PREDATORY BEHAVIOR¹

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Abstract.—A field study of the predatory behavior of *Cerotainia albipilosa* Curran is reported. The asilid foraged from leaves and stems of sunlit plants found along the margins of clearings and paths. The forage flight patterns are discussed. Only 14% of the flights were successful. Most prey (93%) were captured within 2 m of foraging sites. *Cerotainia albipilosa* exhibited specialized methods for capturing prey. The hypopharynx was inserted into the dorsum or one end of the body. The asilid immobilized prey in the air before returning to foraging sites. Individual feeding times averaged 5 minutes but varied considerably depending on prey characteristics and weather conditions. Interfeeding times ranged from 0-35 minutes. While feeding, the asilid frequently hovered in front of a perch and manipulated the prey. Over 94% of the prey belonged to five insect orders (Diptera, Coleoptera, Hymenoptera, Psocoptera and Hemiptera-Homoptera) and 67% to two (Diptera and Hemiptera-Homoptera). Prey were usually soft-bodied, weak flyers and averaged 1.6 mm in length and 0.05 mg in weight. Females captured larger prey than males. Mean predator to prey size and weight ratios were 3.7 and 7.3, respectively. Several factors which influenced prey selectivity are discussed. This asilid was sometimes preyed on by spiders, vespid wasps and other asilids.

In the first part of this study, Scarbrough and Norden (1977) reported on the diurnal activity rhythm and seasonal distribution of *Cerotainia albipilosa* Curran in Maryland. They reported that most of the species flight activities were centered around predation and reproductive behaviors. The purpose of this paper is to report on the predatory behavior of the species. A third paper will deal with reproduction.

Methods and Procedures

General methods and procedures for observations made in the field and the study site were described in a previous paper (Scarbrough and Norden, 1977). Observations were recorded in a notebook at the study site or on a Sanyo solid state tape recorder model 2212 and were later transcribed into a notebook. When possible, photographic records of different behaviors were taken. A Honeywell Pentax 35 mm, single lens reflex camera in conjunction with telephoto lenses and extension tubes was used to obtain close-up pictures.

Collections of prey were made during the summers of 1973 and 1974. Care was taken to collect prey from each segment of the species diurnal activity period through the season to obtain a maximum variety of prey types. Most prey were obtained by capturing asilids seen with prey with a 15 dram snap top vial as I walked through an observation site. The asilid usually ceased feeding immediately and dropped the prey in the vial, although some continued to feed, only to drop them at a later time. This procedure minimized damage to prey specimens. Once the prey was dropped, the asilid was identified and usually released. Some prey were recovered from leaves below feeding sites where asilids had dropped them. The prey were then transferred to vials containing 70% ETOH with the following information: Sex of predator, date, location, and collector.

Prey were analyzed by making two types of body measurements: Size and weight. To determine prey size, each specimen was measured for total body length to the nearest 0.5 mm using an ocular micrometer. From a reference collection made at the study site, prey of selected lengths were oven dried to 38°C for 30 minutes and weighed to the nearest 0.01 mg. Similar measurements of the asilid were obtained by measuring and weighing ten specimens in the same way as described for prey.

Some prey were identified by the author with the aid of the collection housed at the Towson State University Museum. However, most prey were shipped to the Systematic Entomology Laboratory (SEL), IIBIII, Agric. Res. Serv., USDA, Beltsville, Maryland and to the Smithsonian Institution (SI), Washington, D.C. A few specimens were also identified by recognized specialists in other parts of the country.

Results and Discussion

Foraging and feeding.—*Cerotainia albipilosa* foraged under bright skies from leaves and stems of vegetation along margins of clearings and paths, and at air temperatures above 19°C (Scarbrough and Norden, 1977). These flies were usually active on foraging sites, rapidly turning their bodies to face one direction and then another. Each movement was preceded by quick movements of the asilid's head as potential prey flew in front of or to the side of the foraging site. Presumably this behavior enables the asilid to better perceive potential prey and, at the same time, it is in a more suitable position to make a direct forage flight (Dennis and Lavigne, 1975). Similar foraging behavior is exhibited by *Stichopogon trifasciatus* Say (Lavigne and Holland, 1969) and by several species of *Holopogon* (Lehr, 1972; Dennis and Lavigne, 1975).

Most foraging flights were directed toward potential prey in the air after they flew near the asilid. Upon perceiving prey, the asilid "leaped" into the air, flew at an oblique angle to the prey's path, converged and

intercepted it a short distance from the foraging site. Prey were usually captured at the same height or slightly above the foraging site. Adjustments in flight patterns occurred once the asilid took flight. In most cases, after leaving a perch, the asilid's flight path was initially below the prey, although a noticeable elevation in its flight was detected near the interception point. Such behavior facilitated capture, and subsequent immobilization of the prey, in flight. The asilid also compensated for overestimations of flight speed of prey by looping about 5–7 cm above the prey's path, hesitated in this position, and then attacked as the prey passed below at the interception point. They frequently flew toward potential prey only to turn away a few cm from foraging sites or near the interception points. Dennis and Lavigne (1975) termed these flights "investigatory" since they permit discrimination between unsuitable and suitable prey before actually attacking. In a few cases the asilid fluttered its wings and tilted the body forward without leaving a perch in response to passing insects. Unless disturbed, the asilid returned to the same foraging site following each flight.

Females of *C. albipilosa* often flew after insects and falling objects that were several times ($>3\times$) larger than themselves. Several females on separate occasions foraged after butterflies (*Pipilio glaucus* L.) and ichneumonid wasps (*Ophion* sp.), but invariably they turned away from them near the interception points. Two other females attempted to capture a halictid bee (*Augochloropsis* sp.) and a Japanese beetle (*Popillia japonica* Newman). They attacked the insects and held onto their wings for about 1–2 seconds before releasing their grasps and returning to foraging sites. Falling leaves were also frequently pursued by females. Similar observations of asilids flying toward falling objects have been reported by other investigators (Melin, 1923; Parmenter, 1952).

Foraging flights were directed at potential prey flying within a range from 8 cm–4 m in front of or to one side of a perched asilid. Most flights occurred when prey were within 2 m of foraging sites. Flights that resulted in prey captures were made when prey were at the following distances from foraging sites: 68% at 8 cm–1 m, 25% at 1–2 m and 7% at 2–4 m. Ninety percent of the capture flights within 1–20 cm resulted when prey flew from the vegetation and to one side of a perched asilid. Prey captured at greater distances were invariably flying in front of foraging sites. Furthermore, prey captured beyond 2 m by females were characteristically large (>3 mm) whereas those taken at closer ranges represented all size classes. Males captured smaller prey (<2 mm) and were rarely successful ($<1\%$) beyond 2 m. Thus, it is assumed that the visual capability of *C. albipilosa* is more acute at short range.

Few foraging flights resulted in prey capture. Only 14% of the flights were successful regardless of distances from foraging sites. However males



Fig. 1. *Cerotainia albipilosa* feeding on *Aleurochiton* sp. (Hemiptera—Homoptera: Aleyrodidae).

were more successful than females with 17% and 11%, respectively. This forage success differential is directly related to disruptions of the female's foraging behavior caused by courting and mating males (Scarborough and Norden, 1977) and to females foraging longer distances in pursuit of larger prey than males. It is also interesting to note that if these flights were separated into no-contact flights (investigatory) and contact-flights (foraging), as defined by Dennis and Lavigne (1975), then foraging success is much greater (91%). Nevertheless, if one considers the amount of energy which is expended in foraging (investigatory and contact flights), the tendency to fly after most moving objects appears to be quite wasteful. At the same time, since asilids can probably detect only a rough outline of a moving dark body (Melin, 1923), this behavior would maximize the probability that they pursue and capture suitable prey.

Like many asilid species (Dennis and Lavigne, 1975; Shtakel'berg, 1950; Fackler, 1918; Horning and Barr, 1970; Scarborough and Sipes, 1973; Schmid, 1969; Wallis, 1913; Zinov'era, 1959), *C. albipilosa* has specialized methods of capturing prey and common sites for inserting the hypopharynx. The asilid usually captured prey along their dorsolateral surfaces, and impaled them on the dorsum or at one end of the body (Fig. 1). Large bodied insects with hard wings, i.e. the first pair of wings of beetles and cicadellids, were captured with their wings extending in a flight position,

exposing the soft underlying integument. The hypopharynx was inserted into this area, usually near the junction of the thorax and abdomen. Staphylinid beetles and formicine ants were similarly impaled through thin or soft areas of the integument; i.e. eyes and conjunctiva between sclerites at the end of the abdomen. The remaining prey had "soft" integuments, and the hypopharynx was inserted at various places on the dorsum.

Once prey were captured, *C. albipilosa* usually immobilized them in the air before returning to foraging sites. Most immobilizations occurred in flight as soon as they were captured. When prey were large (>2.0 mm), the asilid hovered in front of the foraging site, manipulated them using all six tarsi, and inserted its hypopharynx. Immobilizations were rare events at foraging sites since only one successful and four attempts were observed in over 1,200 observation hours. In these instances, the asilids landed on a leaf, fell on one side, and used all six tarsi to manipulate the prey. The prey were large, and these behaviors were preceded by unsuccessful immobilizations in the air before landing.

Like other asilid species (Dennis and Lavigne, 1975), *C. albipilosa* sometimes foraged with prey impaled upon its hypopharynx. On several occasions the asilids returned with two prey impaled upon their mouth parts, one was the original and the second a new prey. The hypopharynx had been forced completely through the former, and the latter was projecting from its apex. Usually when two prey were captured, the last captured was lost or dropped during manipulation.

Unless disturbed, *C. albipilosa* remained at its initial foraging site until feeding was completed. Males were more "nervous" than females and were easily disturbed. When feeding was completed, the asilid moved its fore tarsi in alternating sequences and disengaged the prey from the hypopharynx. Prey were also discarded by the asilid in flight for another prey, but without using its tarsi. Other investigators (Lavigne and Holland, 1969; Dennis and Lavigne, 1975) have also observed this behavior and have suggested that the hypopharynx is merely retracted into the labrum, allowing gravity to "pull" the prey off the proboscis. While feeding, the asilid continued to observe other moving "prey" which flew near its perch with rapid movements of the head and body.

When feeding, *C. albipilosa* frequently hovered in front of a perch and manipulated the prey. The asilid used all six tarsi to disengage, to rotate and to re-insert the hypopharynx at a new location in the prey. It then returned to the feeding site, re-oriented its body into a foraging position and continued to feed. Prey were sometimes dropped during manipulation in which case the asilid usually returned to the foraging site. However foraging flights were occasionally initiated from the hover position when prey were dropped. The mean time for manipulating prey was 6.1 seconds.

Cerotainia albipilosa fed on prey for an average of 5 minutes. The

length of individual feedings varied considerably, ranging from 1–81 minutes depending upon the size and shape of prey and weather conditions. Aphids such as *Aphis* sp., *Macrosiphum* sp., and *Myzus* sp., which had soft bulbous bodies and average lengths of 2 mm, were fed on for 1–2 minutes. Dipterans and psocopterans, which also had similar integuments and average body lengths, had tubular shapes and were fed on for an average of 3 minutes. Larger prey, such as reproductive ants (*Ponera pennsylvanica* Buckley and *Lasius* sp.) which have hard exoskeletons and constricted “waists,” were fed on for an average of 20 minutes. The asilid frequently manipulated them and alternated the points at which the hypopharynx was inserted from the eyes to the ends of their abdomens. In other large hard bodied prey, such as *Gymnetron pascuorum* (Gyll.) and *Pseudopentarthrum* sp. (Colopetera: Curculionidae) and *Macrosteles* sp. (Homoptera: Cicadellidae), the hypopharynx was alternated from the dorsum below the elytron to the end of the abdomen. Manipulations of aphids, dipterans, psocopterans and other prey with similar body characteristics were rarely observed. Weather conditions frequently influenced the duration of feeding. One asilid fed on *Ponera pennsylvanica* (3.0 mm) for 81 minutes under overcast skies. Feeding on various prey of a similar size under sunlit conditions ranged from 10–20 minutes. Similar observations were reported by Dennis and Lavigne (1975).

The average interfeeding time for *C. albipilosa* was 6 minutes with a range between 0 and 35 minutes. The 0 minute interfeeding time was for individuals who dropped prey and immediately captured new ones. Based upon this data, the average time for a complete feeding and the average 9 hr foraging period, *C. albipilosa* could theoretically feed on a maximum of 49 prey/day. These calculations, like those reported for other asilid species (Lehr, 1964; Musso, 1971; Dennis and Lavigne, 1975, 1976a, 1976b), overestimate the number of prey fed upon by this asilid, since it did not forage and feed continually but often engaged in other activities such as courtship and mating (Scarbrough and Norden, 1977). A more reasonable estimate would be 25–30 prey/day.

Prey.—Most prey of *C. albipilosa*, like other asilid species (Hobby, 1931; Brues, 1946; Poulton, 1906; Cole and Lovett, 1921; Melin, 1923; Dennis and Lavigne, 1975), belong to a few orders of insects. Of the ten orders of prey recorded for this species, over 94% belong to five insect orders and formed 98% of the prey dry weight (Table 1). Prey belonging to Diptera and Hemiptera–Homoptera formed the major prey orders, comprising over 67% of the prey captured and 63% of the prey dry weight. The orders Coleoptera, Hymenoptera and Psocoptera were less significant, forming 27.4% of the total number of prey and 34.1% of the prey dry weight. These results are consistent with prey availability. When sweep samples ($N = 25$) of vegetation were taken at various times at the study sites, Diptera and

Table 1. Dietary composition of *Cerotainia albipilosa* at the Loch Raven Watershed in Baltimore County, Maryland.

Order	No. prey	Percentage prey	Dry weight (mg)	Percentage total dry wt.
Diptera	274	37.1	8.2	24.1
Hemiptera—Homoptera	225	30.4	13.5	39.7
Hymenoptera	88	11.9	3.5	10.3
Coleoptera	60	8.1	4.2	12.3
Psocoptera	55	7.4	3.9	11.5
Others	37	5.0	0.7	2.0
Totals	739		34.0	

Hemiptera–Homoptera were more abundant per sample than other prey orders ($\bar{x}$ /sample; 31.9% Diptera; 39.3% Hemiptera–Homoptera; 19.8% Coleoptera, Hymenoptera, Psocoptera; 9.0% others).

The major prey orders consisted of a wide diversity of species and families (see list of prey). At least 56 and 63 species belonged to 15 and 10 families of Diptera and Hemiptera–Homoptera, respectively. In Coleoptera, Hymenoptera and Psocoptera, the prey consisted of a relatively small number of species (20, 25, 8, respectively) but belonged to several families (12, 13, 7, respectively). It is also interesting to note that within the two major orders, most prey belong to three categories. Over 61% of Diptera belong to Nematocera, and at least 62% of Hemiptera–Homoptera belong to Cicadellidae (22%) and Aphididae (40%). These results were not surprising since the majority of the prey are some of the most abundant plant feeders inhabiting woodland ecosystems.

The average prey captured by *C. albipilosa* was 1.6 mm in length and 0.05 mg in weight (Table 2). The mean predator to prey size and weight ratios were 3.7 and 7.3, respectively. However prey varied considerably ranging from 1–4 mm in length and from 0.01–0.12 mg in weight. Males were rarely found with prey larger than 2 mm in length and 0.09 mg in weight whereas females were found with prey of all class sizes. Most of the prey of males belonging to Diptera, Hemiptera–Homoptera and Hymenoptera consisted of nematocerans, aphids and Apocrita parasitoids, respectively. In addition to the latter prey, females captured a disproportionately greater sample of larger prey, i.e. cicadellids and reproductive formicids, than males.

The tendency of several asilids to capture and feed upon soft-bodied insects is assumed to be directly related to the inability of weak mouth parts to penetrate hard cuticles (Melin, 1923; Martin, 1968; Dennis and Lavigne, 1975). The predaceous behavior and prey records of *C. albipilosa*

Table 2. Mean body lengths and weights of prey arranged according to the sex of the predator.

Orders of prey	Predator sex ^A					
	$\bar{x}$ prey lengths (mm)			$\bar{x}$ prey dry weights (mg)		
	Male	Female	Both sexes	Male	Female	Both sexes
Diptera	1.4	1.5	1.4	0.02	0.03	0.03
Hemiptera—						
Homoptera	1.6	2.0	1.9	0.05	0.07	0.06
Hymenoptera	1.3	2.1	1.8	0.01	0.06	0.04
Coleoptera	1.8	1.7	1.7	0.06	0.07	0.07
Psocoptera	1.4	1.5	1.5	0.06	0.07	0.07
Miscellaneous	1.4	0.7	1.0	0.02	0.02	0.02
Means	1.5	1.7	1.6	0.04	0.05	0.05

^A Predator lengths (mm) 5.6 ♂♂, 6.4 ♀♀; weights (mg) 0.23 ♂♂, 0.43 ♀♀.

support this assumption. Over 73% of the prey were soft-bodied. Among the remaining prey, which included Coleoptera, Hymenoptera, Strepsiptera and Cicadellidae, the predator utilized specific techniques to capture and to immobilize them. The hypopharynx was invariably inserted in areas where the cuticle was thin or soft.

Other investigators (Lehr, 1958; Hobby, 1931; Dennis and Lavigne, 1975) have suggested that differences in predatory habits of the two sexes were the result of temporal segregation of activity patterns, and differential densities and nutrition requirements of females. This study supports these suggestions. Males of *C. albipilosa* spend less time foraging and feeding in afternoons, and more time searching for and mating with females (Scarborough and Norden, 1977). Females foraged throughout the day, and thus were found more frequently with prey than males. Furthermore several prey types captured by females were active and abundant at times when males were involved in other activities. Females captured most of the coleopterans and psocopterans between 1:00 and 4:00 PM. Flying reproductive ants swarmed and were captured during hot humid afternoons which were preceded by rain. Females, which had well-developed ovaries, evidenced by the presence of numerous eggs, weighed much more than an equal number of males ($N = 20$, $\bar{x} = 0.43$ mg ♀♀, 0.23 mg ♂♂). However, females whose ovaries and eggs were removed by dissection, weighed about the same ($N = 20$, $\bar{x} = 0.28$ mg) as males. Thus the difference in body weights of the sexes is indirectly related to differences in predatory habits. Females must spend more time foraging and feeding to obtain additional nutrition for the continuous production of eggs.

Movement of prey evokes feeding behavior in numerous predatory animals, provided that the object falls within certain size limits (Marler and Hamilton, 1956). Melin (1923) suggested that the vision of asilids is not well developed and that they perceive prey as dark moving objects. *Cerotainia albipilosa* appears to be "programmed" to forage after almost any small moving object that passes near its perch. Motion of insects in flight and falling leaves were sufficient to stimulate the asilid to leave a perch in pursuit. All prey, except spiders, were winged and captured in flight. The spiders were immature, and in effect "flying" since they were either "ballooning" or moving at the ends of suspended silk threads. Wings of most prey were large and extended to or beyond their bodies. Furthermore, prey moving in front of a perched asilid interrupt the rays of light, casting a shadow upon itself. The fluttery motion produced by large wings of soft-bodied prey, together with a dark shadowed body, greatly increase the total size of the prey's body, and undoubtedly form the major cues which stimulate the asilid to leave its perch in pursuit.

A few asilid species are apparently capable of utilizing color to select prey (Linsley, 1960; Bohart, 1958). Dennis et al. (1975) showed that when *Efferia frewingi* Wilcox was presented with black, orange and white models of various sizes and shapes, the asilid responded preferentially to black, oblong ones. However use of colors to select prey by *C. albipilosa* was not detected. Color of prey integuments varied considerably, ranging from light yellow to dark brown. Color detection would be difficult at best and probably could not occur until the predator was about to, or was in contact with the prey because 1) the flight path of the asilid is slightly below that of the prey until the two arrive at the interception point, and 2) the asilid approaches the prey on its shadowed side.

The following is a list of prey taken by *C. albipilosa* at the Lock Raven Watershed, in Baltimore County, Maryland. All prey were collected at the study site between 30 June and 30 August of 1973 and 1974. In some instances prey is presented only to the order or family level since specific identifications are not yet available. Each notation of prey refers to a single record in the list of prey unless followed by a number in parentheses. All prey are adults except for Araneida. W. B. Peck (Araneida); D. R. Smith (Isoptera); E. L. Mockford (Psocoptera); K. O'Neill (Thysanoptera); J. L. Herring, L. M. Russell, J. P. Kramer, R. C. Froeschner (Hemiptera-Homoptera); C. W. Sabrosky, W. W. Wirth, R. J. Gagné, G. C. Steyskal, L. Knutson (Diptera); A. S. Menke, B. D. Burks, P. M. Marsh, D. R. Smith (Hymenoptera); D. M. Anderson, R. E. Warner, R. E. White, J. M. Kingsolver, P. J. Spangler (Coleoptera); D. R. Davis (Lepidoptera); are thanked for the identification of their respective groups. Froeschner, Spangler and Peck are with the Smithsonian Institution.

Prey captured by *C. albipilosa*. ARANEIDA, Unidentified (1) 31.VII.74;

Theridiidae, *Oecobius* sp. 7.VII.73; Linyphiidae (2) 31.VIII.74; Araneidae (2) 31.VII.74; Agelenidae (3) 30.VII.74, 1.VIII.74, 3.VIII.74; Clubionidae, Clubioninae (3) 31.VII.74; Thomisidae, Philodrominae (2) 31.VII.74, 12.VIII.74; Salticidae (3) 10.VII.74, 16.VII.74, 17.VII.74; ISOPTERA, Rhinotermitidae, *Reticulitermes flavipes* (Kollar) (6 winged reproductives) 1.VII.73; PSOCOPTERA, Amphipsocidae, *Polypsocus corruptus* (Hagen) 16.VII.74, 30.VII.74; Caeciliidae, *Caecilius aurantiacus* (Hagen) 19.VII.74, 31.VII.74, 6.VIII.74; Ectopsocidae, *Ectopsocopsis cryptomeriae* (Endln.) (2) 20.VII.73, (7) 12.VII.74, (9) 17.VII.74, (2) 22.VII.74, (3) 1.VIII.74, (2) 3.VIII.74; Lachesillidae, *Lachesilla pallida* (Chapman) (1) 20.VII.74; Peripsocidae, *Peripsocus quadrifasciatus* (Harris) (1) 1.VII.73; *P. alboguttatus* group (1 sp.) 30.VI.74; Philotarsidae, *Aaronella* sp. (2) 15.VII.74, (2) 19.VII.74, (7) 31.VII.74; Psocidae, *Trichadenotecnum alexandrae* Somm. 19.VII.73, 2.VIII.73, 19.VII.74; Unidentified (2) 16.VII.74, 31.VII.74, 2.VIII.74, 15.VIII.74; THYSANOPTERA, Aeolothripidae, *Acolothrips vittipennis* Hood (2) 9.VII.74, Thripidae *Frankliniella tritili* (Fitch) 10.VII.74, *F. runneri* (Morgan) 12.VII.74, *Anaphothrips obscurus* (Mueller) 15.VII.74, *Limothrips cerealium* (Haliday) 16.VII.74, *Chaetanaphothrips* sp. near or = *orchidii* (Moulton) 17.VII.74, Unidentified 19.VII.74, Phlaeothripidae, *Leptothrips* sp. near or = *mali* (Fitch) (3) 31.VII.74, *Hoplothrips fieldsi* Crawford (2) 1.VIII.74, *Liothrips* sp. 1.VIII.74; HOMOPTERA-HEMIPTERA, Aleyrodidae, Aleyrodinae 22.VII.74, 31.VII.74, 1.VIII.74, *Aleurochiton* sp. (?) 3.VII.73; Aphididae, *Amphoraphora* sp. (2) 8.VII.73, *A. sensoriata* Mason 7.VII.73, *Anoecia cornia* (F.) (3) 30.VII.74, *A. querci* (Fitch) 3.VIII.74, (2) 15.VIII.74, *Aphis* sp. 27.VII.73, 1.VIII.74, *A. gossypii* Glov. 27.VIII.74; *A. rubifolii* (Thos.) 29.VII.74, *A. sambucifoliae* Fitch 12.VII.74, *Capitophorus elaeagni* (Del Guer.) 19.VII.74, 22.VII.74, 1.VIII.74, *Chaitophorus* sp. 1.VIII.74; *C. hippophaes* (Wik.) 17.VII.74, 3.VIII.74, *C. pusillus* Hottes and Frison 27.VII.74, Drepanosiphinae 30.VII.73, *Drepanaphis* sp. 25.VII.73, *D. acerifoliae* (Thos.) 29.VII.74, *D. saccharini* Smith and Dillery 6.VIII.74, *Dysaphis* sp. 16.VII.74, *D. radicola* (Mord.) 31.VII.74, *Essigella pini* Wilson 22.VIII.74, *Hyadaphis foeniculi* (Pass.) 1.VIII.74, *Hyalopterus pruni* (Geof.) 15.VIII.74, *Macrosiphon* sp. 30.VI.74, *M. liriodendri* (Monell) (3) 30.VI.74, 12.VII.74, 16.VII.74, 19.VII.74, (2) 30.VII.74, *M. avenae* (F.) 30.VI.74, 16.VII.74, 31.VII.74, 1.VIII.74, *M. euphorbiae* (Thomas) 1.VIII.74, *Mastopoda pteridis* Oestlund 27.VIII.74 *Monellia* sp. 30.VI.74, *M. costalis* (Fitch) 1.VII.73, *Monelliopsis* sp. (2) 19.VII.74, 31.VII.74, 20.VIII.74, *Myzocallis* sp. (2) 1.VIII.74, 15.VIII.74, (2) 23.VIII.74, *M. asclepiadis* (Monell) (2) 1.VIII.74, 15.VIII.74, (2) 22.VIII.74, 23.VIII.74, *Myzus persicae* (Sulz.) 1.VIII.74, *Masonovia* sp. 7.VIII.74, *Ovatus phyloxae* (Sampson) 12.VII.74, *Prociphilus fraxinifolii* (Riley) 9.VII.74, 15.VII.74, 18.VII.74, 29.VII.74, *Rhopalosiphum* sp. 1.VIII.74, *R. maidis* (Fitch) 16.VII.74, 19.VII.74, 22.VII.74, 30.VII.74, 31.VII.74, 1.VIII.74, *Schizolachnus* sp. 16.VII.73, *Thecabius*

sp. 12.VII.74, *Therioaphis trifolii* (Thos.) 15.VII.74, *Tinocallis kahawaluokalani* (Kirk.) 17.VII.74, 19.VII.74, *T. ulmifolii* (Monell) 7.VII.74, 9.VII.74, 12.VII.74, 16.VII.74, 17.VII.74, 19.VII.74, 22.VII.74; Unidentified 30.VI.74, (2) 12.VII.74, (4) 17.VII.74, (2) 18.VII.74, (4) 19.VII.74, 27.VII.74, (2) 29.VII.74, (3) 31.VII.74, (5) 1.VIII.74, 15.VIII.74, 22.VIII.74; Cicadellidae, *Agallia constricta* Van Duzee 16.VII.74, *Alebra albostrigella* (Fallen) (2) 22.VII.74, *Aphrodes* sp. 1.VII.73, 7.VII.73, (4) 8.VII.73, 16.VII.73, 10.VII.74, 19.VII.74, *Balclutha* sp. (2) 7.VII.73, *Coelidia olitoria* (Say) 20.VII.74, Deltocephalinae 23.VII.73, *Dikraneura* sp. 7.VII.73, 12.VII.74, 15.VII.74, 18.VII.74, *D. mali* (Provancher) 16.VII.74, 1.VIII.74, *Empoasca* sp. 16.VII.74, 17.VII.74, 20.VII.74, *E. bifurcata* DeLong (2) 25.VII.73, *Erythro-neura* sp. 7.VII.73, 8.VII.73, (2) 12.VII.74, 15.VII.74, (3) 16.VII.74, (2) 20.VII.74, 29.VII.74, (2) 31.VII.74, *E. tricineta* Fitch 14.VII.74, *E. vulnerata* Fitch 12.VII.73, *Forcipata loca* DeLong and Cardwell 20.VII.73, 1.VIII.74, *Graminella nigrifrons* (Forbes) (2) 29.VII.73, 14.VII.74, 30.VII.74, 31.VII.74, 1.VIII.74, 3.VIII.74, *Macrosteles fascifrons* (Stål.) 8.VII.73, 10.VII.73, 20.VII.74, 30.VII.74, *M. slossoni* (Van Duzee) 12.VII.73, *Onecpsis verticis* (Say) 30.VII.74, 2.VIII.74, *Scaphytopius* sp. 30.VII.74, *S. acutus* (Say) 31.VII.74, *S. amplus* DeLong and Mohr 3.VIII.74, Typhlocybinae 30.VII.74, 31.VII.74, *Xestocephalus pulicarius* Van Duzee 30.VII.74; Cixidae 20.VII.74; Delphacidae, *Delphacodes* sp. 16.VII.73, *D. puella* Van Duzee (2) 27.VII.74, *Pissonotus* sp. 22.VII.74; Derbidae, *Cedusa gedusa* McAtee 29.VII.74; Miridae, *Lygus lineolaris* (P. daB.) 7.VII.73, Unidentified (2) 15.VII.74, (3) 16.VII.74, (3) 29.VII.74, (4) 31.VII.74, (3) 1.VIII.74, (5) 15.VII.74, (4) 23.VIII.74; Phylloxeridae, *Phylloxera* (3) 15.VIII.74, Psyllidae, *Craspedolepta* sp. 16.VII.74, *C. fumida* Caldwell 16.VII.74, *Psylla annulatus* Fitch 1.VIII.73, Unidentified 7.VII.73, 8.VII.73; Tingidae, *Corythucha arcuata* (Say) 29.VII.74, (2) 3.VIII.74, LEPIDOPTERA, Tineidae 31.VII.74, Unidentified (2) 1.VIII.74; DIPTERA, Unidentified 16.VII.74, (2) 30.VII.74, (13) 31.VII.74, (13) 1.VIII.74, (2) 12.VIII.74, 15.VIII.74, (3) 23.VIII.74; Agromyzidae, *Cerodontha dorsalis* (Loew) 29.VII.74, *C. (Poemyza) muscina* (Meigen) 17.VII.74; Cecidomyiidae, Cecidomyiidi 9.VII.74, 10.VII.74, (2) 12.VII.74, (3) 20.VII.74, 1.VII.74, *Anarete* sp. 29.VII.73, 27.VII.74, 2.VIII.74, 3.VIII.74, (2) 20.VIII.74, *A. pritchardi* Kim 20.VII.73, *Asteromyia* sp. 12.VII.74, *Atrichopogon levis* (Coq.) 20.VII.74, *Contarinia* sp. 19.VII.74, *Culicoides paraensis* (Goeldi) 20.VII.74, 22.VII.74, *Dasineura* sp. 17.VII.74, 22.VII.74, 27.VII.74, (2) 31.VII.74, *Dasyhelea* sp. 22.VII.74, 30.VII.74, 31.VII.74, 3.VIII.74, *Forcipomyia* sp. 30.VI.74, *F. brevipennis* (Macq.) 30.VI.74, *Hyperdiplosis* sp. 15.VII.74, *Lasioptera* sp. (2) 16.VII.74, *Lestremia* sp. (3) 10.VII.73, 28.VII.73, *Lestodiplosis* sp. 12.VII.74, *Micromyia* sp. 9.VII.73, 15.VII.73, 18.VII.73, 25.VII.73, (2) 17.VII.74, *Neolosioptera* 7.VII.73, 12.VII.74, 19.VII.74, *Procystiphora* sp. 30.VI.74, 7.VII.74, 19.VII.74, *Porricondyla* sp. 19.VII.74, 31.VII.74, *Resseliella* sp. 30.VII.73, (2) 16.VII.74, (2) 17.VII.74,

22.VII.74, 3.VIII.74; Chaoboridae, Unidentified (3) 7.VII.73, *Chaoborus* sp. (2) 30.VI.74; Chironomidae, Orthoclaadiinae (3) 1.VII.73, 30.VII.74, (2) 7.VII.74, (3) 12.VII.74, (4) 15.VII.74, 18.VII.74, (2) 19.VII.74, 20.VII.74, (3) 22.VII.74, (5) 31.VII.74, 2.VIII.74, 6.VIII.74 (3) 15.VIII.74, (2) 23.VIII.74, *Anatopynia* sp. 22.VII.74, *A. dyari* (Coq.) 10.VII.73, *Chironomus* sp. 12.VII.74, 17.VII.74, 20.VII.74, *Cricotopus* sp. (2) 20.VII.73, 25.VII.73, (2) 26.VII.73, (2) 16.VII.74, 22.VII.74, *Procladius bellus* (Lw.) 16.VII.74, *P. culiciformis* (L.) 18.VII.73, *Tanytarsus* sp. 30.VII.74; Chloropidae, *Conioscinella* sp. 16.VII.74, *Elachiptera umbrosa* (Lw.) 7.VII.73, *Goniopsita catalpae* (Mall.) 1.VIII.74, *Oscinella carbonaria* (Lw.) 25.VII.73, 29.VII.73, (2) 31.VII.74, *O. painteri* Sabr. 28.VII.73, *Siphonella nigripalpis* (Mall.) 30.VII.74, *Thaumatomyia bistrata* (Wlk.) 2.VIII.74, *T. glabra* (Mg.) 31.VII.74; Dolichopodidae, *Chrysotus* sp. (5) 1.VII.73, 7.VII.74, 12.VII.74, 18.VII.74, *Gymnopternus debilis* Loew 17.VII.73, *Thrypticus* sp. 7.VII.73, Drosophilidae, *Scaptomyza adusta* (Lw.) 12.VII.74, *S. pallida* (Zett.) (5) 7.VII.73, 15.VII.74, 18.VII.74, 20.VII.74, 30.VII.74, 6.VIII.74, *S. wheeleri* Hackmann 30.VII.74; Lonchopteridae, *Lonchoptera furcata* (Fallen) 1.VIII.74; Phoridae, Unidentified (2) 16.VII.73, (2) 15.VII.73, *Megaselia* sp. (3) 7.VII.73, 7.VII.74, 12.VII.74, 15.VII.74, (3) 16.VII.74, (4) 17.VII.74, (3) 31.VII.74, *Puliciphora* sp. (6) 16.VII.74, (2) 17.VII.74, (4) 22.VII.74, (2) 12.VIII.74; Psychodidae, Unidentified 28.VII.73; *Psychoda* sp. (2) 1.VIII.74; Scatopsidae, Unidentified 8.VII.73, *Scatopse fuscipes* Mq. 31.VII.74, *Rhegmoclema* sp. 31.VII.74, 6.VIII.74, 18.VII.74; Sciariidae, *Bradysia* sp. (3) 1.VII.73, (4) 12.VII.74, (2) 15.VII.74, (4) 16.VII.74, (2) 20.VII.74; Sepsidae, *Sepsis punctum* (Fab.) 12.VII.74; Sphaeroceridae, *Leptocera* sp. (5) 16.VII.73, (2) 22.VII.73, 20.VII.74, (2) 30.VII.74, 31.VII.74, *L. (Pterogramma) palliceps* Johnson 16.VII.74, *Sphaerocera pusilla* (Fallen) 7.VII.73, *S. vaporarium* Holaday 31.VII.74, 1.VIII.74; Stratiomyidae, *Microchrysa polita* (L.) (2) 31.VII.74, 1.VIII.74, *Oxycera* sp. 15.VIII.74; Tipulidae, 16.VII.74: COLEOPTERA, Unidentified 30.VI.74, (2) 12.VII.74, 22.VII.74, (3) 31.VII.74, Alleculidae, *Mycetochara haldemani*: LeC. 2.VIII.74, Chrysomelidae, *Baliosus* sp., 12.VII.74, 16.VII.74; 20.VII.74, 25.VII.74, *Chaetocnema* sp. 25.VII.73, 30.VII.74, (2) 31.VII.74, (2) 1.VIII.74, 11.VIII.74, 14.VIII.74, 25.VIII.74; Curculionidae, *Gymnetron pascuorum* (Gyll.) 16.VII.73, 17.VII.74, 19.VII.74, *Microtrogus picirostris* (F.) (3) 17.VII.74, *Pseudopentarthrum* sp. 25.VII.73, 1.VIII.73; Hydrophilidae, *Cercyon* sp. 25.VIII.74, 27.VIII.74; Lathrididae, *Corticaria* sp. 27.VIII.74; Leptodiridae, *Nemadus* sp. prob. *parasitus* LeC. 22.VIII.74; Leiodidae, *Colenis impunctata* LeC. 28.VIII.74; Mycetophagidae, *Litargus quadrispilatus* LeC. 31.VII.74, *L. nebulosus* LeC. 31.VII.74; Phalacridae, *Stilbus* sp. (2) 31.VII.74 (2) 1.VIII.74, 2.VIII.74, *Olibrus* sp. 28.VII.74; Ptilodactylidae, *Ptilodactyla angustata* (3) 16.VII.73; Rhipiphoridae, *Rhipiphorus* sp. 18.VII.74, *Hypothenemus* sp. (3) 9.VII.74, *Pityophthorus* sp. 7.VII.73, *Pityogenes hopkinsis* Swaine 20.VII.74; Staphylinidae, Unidentified, (2) 15.VII.74, (5) 16.VII.

74, 22.VIII.74, 23.VIII.74: STREPSIPTERA, Stylopidae, *Pseudoxenos lugubris* (Pierce) 18.VII.74, Unidentified 31.VII.74: HYMENOPTERA, Unidentified (2) 16.VII.74, 19.VII.74, (3) 31.VII.74, Aphelinidae, Unidentified 27.VII.74, 1.VIII.74; Aphidiidae, *Aphidius* sp. 18.VII.74, *Lysiphlebus* sp. 30.VII.74; Brachonidae, *Asobara* sp. 16.VII.74, 19.VII.74, 30.VII.74, *Aspilota* sp. 20.VII.74, 31.VII.74, *Chorebus* sp. 2.VIII.73, *Oenonogaster* sp. 12.VIII.73, *Synaldis* sp. 20.VII.74, 31.VII.74; Ceraphronidae, *Ceraphron* sp. (4) 17.VII.74, *Conostigmus* sp. (2) 31.VII.74, *Lygocerus* sp. 12.VII.74, (2) 16.VII.74, 30.VII.74; Chalcididae, *Euchrysa* sp. 25.VII.73; Cynipidae, Encoilinae 16.VIII.74, *Alloxysta* sp. 3.VIII.74; Dryinidae, Unidentified 31.VII.74, 1.VIII.74, Encyrtidae, new *Encyrtus* sp. 10.VII.74, 12.VII.74, 16.VII.74, 19.VII.74, 31.VII.74, 1.VIII.74; Eulophidae, Unidentified 31.VII.74; Eurytomidae, Unidentified 16.VII.74; Formicidae, *Dolichoderus* sp. 16.VII.74, *Lasius* sp. (10) 26.VII.73, (2) 22.VII.74, (3) 27.VII.74, 15.VIII.74, *Leptothorax* sp. (3) 28.VII.74, *Monomorium minimum* (Buckley) 30.VI.74, *Paratrechina* sp. 26.VII.73, 19.VII.74, *Ponera pennsylvanica* Buckley 31.VIII.74, (4) 1.VIII.74, (5) 23.VIII.74, (3) 27.VIII.74, *Solenopsis molesta* (Say) 1.VIII.74; Mymaridae, Unidentified 12.VII.74, 16.VII.74; Pteromalidae, Pteromalini 16.VII.74, 19.VII.74, (2) 31.VII.74.

Enemies.—*Cerotainia albipilosa* are sometimes preyed on by arthropods larger than themselves. Thirteen instances of predation were observed: *Theridon* sp. (Tetragnathidae: Araneida), *Vespula* sp. (2) and *V. arenaria* (F.) (8) (Vespidae: Hymenoptera), *Efferia aestuans* (L.) and *Dioctria tibialis* McAtee (Asilidae: Diptera). Several unsuccessful attacks were directed by the following: *Pisaurina mira* (Walkenuer) (Pisauridae: Araneida), *Misunera* sp. (Thomopsidae: Araneida), *Laphria sicula* McAtee, *D. tibialis* (Asilidae: Diptera), and *Vespula* spp. Each predator used a specific attack strategy. *Vespula* spp. flew along the margin of the study site and hovered frequently in front of occupied perches. This behavior "flushed" the asilid which was attacked as it attempted to escape. *Vespula* spp. sometimes attacked before the asilid attempted to fly away. *Theridon* sp. captured its prey with a web while the other araneids attacked from a concealed position below a foraging site. The asilid predators, which were usually perched near the prey, attacked *C. albipilosa* when it flew from a perch. While cannibalism by this species was not observed, males and females displayed agonistic behavior toward other males (Scarborough and Norden, 1977).

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Footnote

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